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Burrowing Nematode on Anthurium: Recognizing Symptoms, Understanding the Pathogen, and Preventing Disease

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Burrowing nematodes (species of *Radopholus*) are common in subtropical and tropical regions and are known to attack over 600 plant species. Severe economic damage to citrus is caused by *Radopholus similis*, and *R. oryzae* is a serious pathogen of rice. In Hawaii, burrowing nematodes have been associated with roots and rhizomes of banana, roots of coffee, and roots or stems of aglaonema, calathea, dieffenbachia, dracaena, potted palms, and other ornamentals[3].

R. similis causes major problems in banana, infecting roots and rhizomes[2]. Infected banana plants with weakened root systems are stressed during fruit production and are often uprooted during storms. The nematodes feed on the cortical cells of fleshy roots, blackening and killing the roots. Widespread distribution of this nematode throughout the tropics has resulted from use of infested planting material to establish new banana fields.

Some isolates of *R. similis* are highly destructive on citrus and cause root rots that lead to tree decline[2]. Nematodes actively feed on the tips of citrus roots and prevent normal root growth. High populations of nematodes feeding on the plant cause the leaves to turn yellow and decrease in size, resulting in fewer, smaller fruits. Eradication of nematodes from citrus orchards is costly and difficult, so prevention of field infestation is of critical importance.

R. similis also causes root and stem rots in anthurium[1], but the phases of nematode penetration and disease development in anthurium are not well known or widely recognized. In this publication we describe symptoms that are clues to the presence of burrowing nematodes in anthurium. It is particularly important to look for these symptoms when older plantings are used as stock plants for new fields, to prevent introduction of the nematodes at planting. The biology of *Radopholus* species is also described, and disease prevention measures are discussed.

> Crops susceptible to attack by burrowing nematodes include alfalfa, anthurium, avocado, bamboo, banana, bean, beet, bird-of-paradise, cabbage, carrot, castor bean, citrus, coconut, coffee, eggplant, ginger, guava, lettuce, mango, onion, pepper, rice, tomato, and many others.

Disease symptoms and nematode movement

Typical symptoms

Root rots and plant decline are well known symptoms of burrowing nematode infection. Anthurium root rots caused by *R. similis* are brown or dark brown to black. The rots develop relatively slowly. Initially, although older roots are infected and rot, new roots are produced and the plant often continues to grow well. But compared to healthy plants (Fig. 1), the amount of functional roots in diseased plants is greatly reduced. With time, fewer new roots are produced, and gradually the entire root mass is destroyed (Fig. 2). The steady and progressive destruction of the root system usually causes plant decline in the second to fourth years. The leaves yellow and may have other symptoms of nutrient deficiency. The plants become smaller and lose vigor, producing fewer and smaller flowers.

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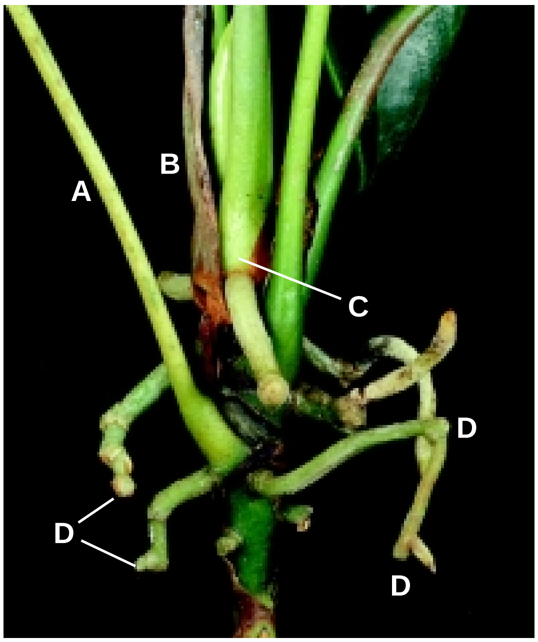
1. Healthy young anthurium plant with a good root system.

2. Typical root rots caused by the burrowing nematode on several anthurium plants. Note the extensive, brown root rots, yellow leaves, and stunted plants.

3. Anthurium root tip infected with burrowing nematodes.



4. Typical stubby roots on an anthurium plant: A, petiole; B, sheath; C, node; D, stubby roots.

Unrecognized symptoms

Stubby root

In moist environments, the nematodes are able to migrate short distances outside the plant, above the soil line. They maybe splashed onto or migrate up stems to aerial roots, where they penetrate the soft, fleshy root tips. Invasion of the root tips frequently cause cessation of root growth. Infected tips are brown (Fig. 3) and may be missing.

When the environment is dry, nematodes either migrate back into the soil or desiccate, and many rotted tips are devoid of nematodes. The tips produce a callus, stop growing, and have a rounded form that suggests the description “stubby root.” When the environment becomes favorable for root growth, a new lateral root is produced at or near the tip of the original root. This new tip can be infected and become stubby also. Repetition of this cycle results in a short aerial root with several stubby tips (Fig. 4).

Stubby roots and branched aerial roots are not normal in anthurium. While this condition is not always caused by nematodes, these abnormal roots should be a clue to the possible presence of burrowing nematodes.

5. Small, light brown rots are early stages of nematode penetration into anthurium roots.

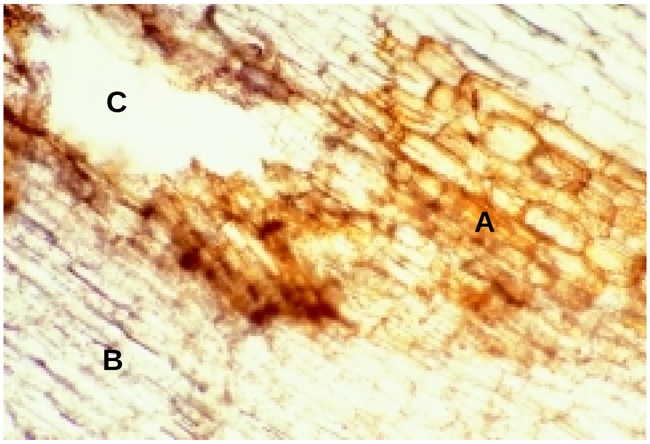
6. Expanding root lesions with yellow borders are infected with burrowing nematodes.

Small spots on roots

The first symptom of rot on roots growing in cinder potting medium are small, pink, orange-brown or light brown streaks or elongated rots (Fig. 5). These develop into brownish streaks, with or without yellow borders (Fig. 6), and then become distinctly long, blackish-brown rots (Fig. 7). With time, these rots expand, and large sections of the root are destroyed (Fig. 8). At times, if the nematodes feed and grow deep into the root, only a gray streak is visible on the external surface of a white or light colored root.

7. Expanding lesion (spot) on an anthurium root.

8. Early stages of root rot on anthurium. Note the missing root tips with exposed vascular strands.



9. A thin slice of a small spot on an anthurium root was prepared and photographed under a microscope: A, a region of dead or damaged cells caused by burrowing nematodes; B, healthy cells surrounding the damaged area; C, cavity formed by the activity of nematodes and other microorganisms.

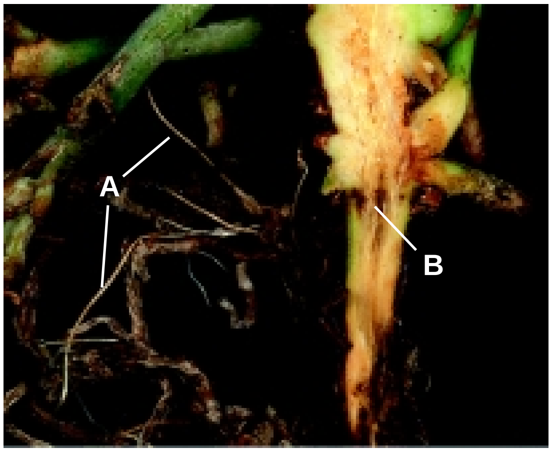
11. Photomicrograph of a female burrowing nematode: A, the head and stylet; B, the vulva or opening from which eggs are laid; C, the tail.

10. Photomicrograph of a burrowing nematode within a small anthurium root cavity. About half of the nematode body is shown. Its head and stylet are within the circle.

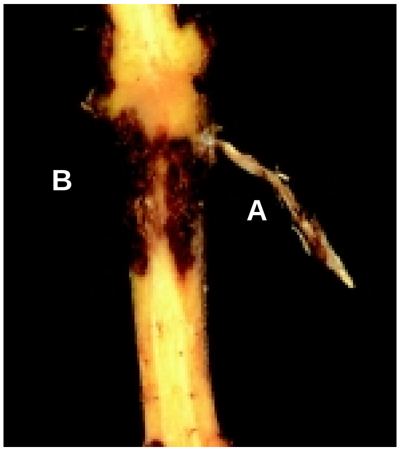
12. Photomicrograph of a nematode egg (high magnification).

Burrowing nematodes are easy to find within these small rots (Figs. 9, 10, and 11). Each streak has 30–60 nematodes, with larger lesions containing eggs (Fig. 12) and juvenile nematodes. With time, the entire section of the root becomes brown as secondary organisms (including nonpathogenic plant nematodes, fungi, protozoa, bacteria, and others) invade the rot and cause complete decomposition of the internal root tissue. Weak fungal pathogens may also increase the rate of decay. Dead roots consist of a hollow brown tube (the original epidermal layer) around a fibrous core, the remains of the vascular system formerly used to transport water and nutrients (Fig. 13). Completely rotted roots have few nematodes, as the burrowing nematode feeds on living plant tissue.

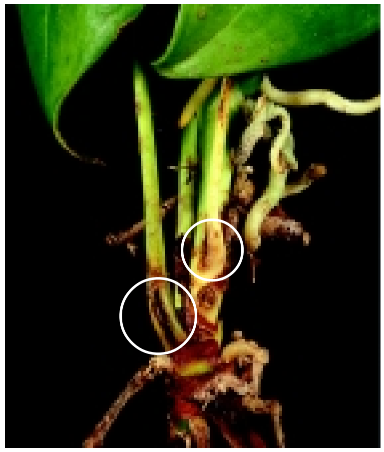
New roots are produced from the stem near a node, where the leaf stem (petiole) attaches to the plant stem. Infected roots thus provide nematodes entry into the stem (Figs. 13 and 14).



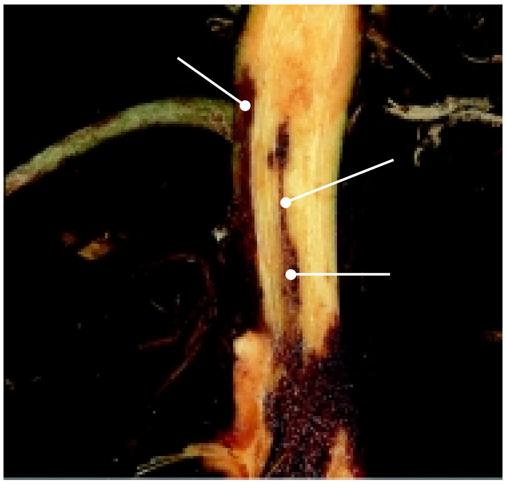
13. Severely diseased roots with vascular fibers exposed. A, fibers; B, stem rot.



14. Stem rots initiated from root rots on anthurium. A, rotted root; B, stem rot.



15. Hidden petiole rots were exposed by removing the brown sheath that covers the lower petiole.



16. Progression of burrowing nematode damage from older stem rots into healthier stem areas as indicated by the elongated rot symptom.

17. (Above and right) Typical white or light colored vascular fibers in rots of mature anthurium stems.

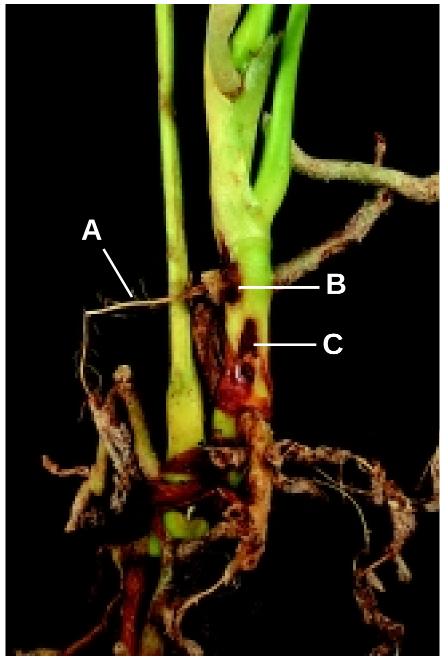
Petiole and sheath infections

In many cultivars of anthurium a small, moist cavity is often present at the nodes where petioles attach to the stem, and these cavities harbor nematodes. The tender, tiny bud at the node provides soft tissue through which burrowing nematodes enter the plant, feed, grow, and reproduce. The sheath that covers the base of the petiole at the node also creates a conducive environment for nematodes, and infection of the petiole base is common (Fig. 15). Infections of the petiole also lead to stem infections.

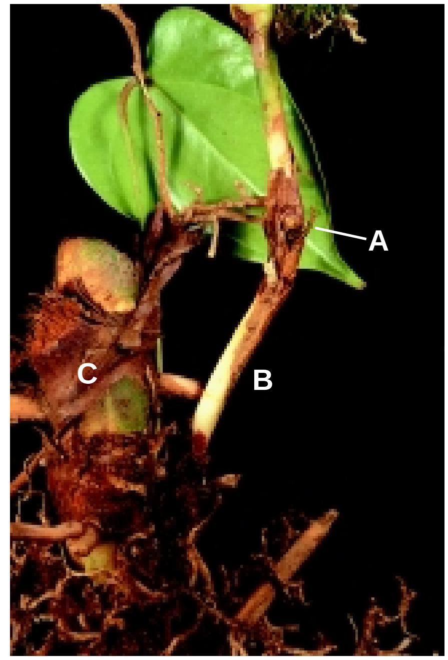
White fibers in stem rots

Typical rots of the main stem are brown to black on the outside and are generally started from diseased roots or nematodes on the original planting material. Within the stem, nematodes feed and move progressively up the stem from the older stem rots (Fig. 16)[6]. Internally, stem rots of the older stem tissue are generally dry, brown, and crumbly, with white to off-white fibers of the vascular system (Fig. 17). Nematodes cannot be seen without a microscope, but the symptoms they cause allow

18. Stem tip destroyed by burrowing nematodes. Field symptoms were death of the youngest leaf and soft rot of the tip of the plant. A, petiole or leaf stem of the youngest leaf; B, diseased sheath; C, dead tip associated with very high numbers of burrowing nematodes.



19. Infection of a young stem following root infection. A, root rot; B, stem rot; C, stem rot from the root rot below.



20. Young stem infected from root rot. A, root rot; B, stem rot; C, mature stem of original plant.

us to follow their movement and detect their presence.

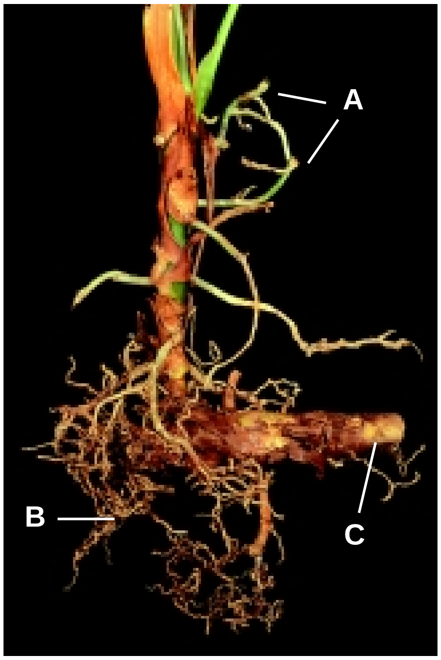
In contrast, mature stems infected with the bacterial blight pathogen *Xanthomonas* generally have black vascular streaks surrounded by white to off-white cortical cells. Invasion of the vascular system by *Xanthomonas* kills and discolors the vascular cells, forming dark lines within the stem. High numbers of bacteria are found within these streaks.

These symptoms, although valuable for diagnosis in older stems, can be confusing in young tissue or near the tip of the plant, where stem tissue is soft, moist, and not fibrous. In one of our investigations, a single, pin-sized, dark line was traced from an active nematode rot in the older section of a stem to its apical tip, where burrowing nematodes were found. In another study, burrowing nematodes were extracted 2½ inches above the base of the stem in plants about 12 inches tall[6], and they were also extracted from petioles. Thus nematodes may be fairly common in stem sections without obvious symptoms.

Rots of the stem tip

Stem tip rots are soft, moist, and brown, without white fibers. These apical rots are generally attributed to bacterial blight. However, burrowing nematodes and the tomato spotted wilt virus[4] can also cause tip rots. High numbers of burrowing nematodes have been found in these apical rots (Fig. 18). If a field has been free of bacterial blight and this symptom occurs, a sample should immediately be sent to the CTAHR Agricultural Diagnostic Service Center to determine the cause. Any large population of nematodes in the plant canopy will quickly spread to neighboring plants by splashing water. Tip blights must be promptly removed from the field regardless of the causal agent (that is, *Xanthomonas* or burrowing nematode). The need for other control measures and the suitability of the field for future use as a stock source will depend on the particular causal agent.

Young shoots (*keiki*) produced by older plants are prime targets for burrowing nematodes. These young plants are produced at the base of older plants, near the



21. Young plant produced from a contaminated mature stem piece (gobo). Symptoms of nematode infestation and infection are A, stubby roots; B, numerous rotted roots; C, mature stem piece of original plant.

22. Above-ground stem section that externally appeared healthy; after the stem was split, nematode damage was revealed. A, stubby roots; B, active internal stem rot.

soil line. New roots and immature shoot tissues are easily invaded by nematodes, and typical brown lesions form (Figs. 19, 20).

Infected gobo (propagative stem cuttings)

When "gobo" (stem cuttings) are used to produce new plants, a surprising number are contaminated with burrowing nematodes. In fact, we find that contaminated cuttings are more common than clean cuttings at some nurseries. This may be because growers do not realize that the nematodes are present in stem pieces above ground. The nematode symptoms are hidden beneath sheaths in moist crevices as small petiole rots, on aerial roots, or as pin-point internal stem infections (Fig. 21). Research has shown that plants with roots free of nematodes have had stems infected with nematodes[6].

Some brown stem rots may be assumed to be fungal infections. However, in our recent isolations of pathogens from anthurium tissues, almost all have yielded nematodes, while common pathogens such as *Pythium* were not isolated. Thus we observe that the burrowing nematode problem is returning to the industry as stem cuttings become an increasingly common means for propagation (Fig. 22).

Stem cuttings produce a new plant in a shorter period of time and at a lower cost than tissue-cultured plants. Gobos can produce plants that are healthy and vigorous, but they must be uncontaminated when planted. Growers often wonder where the burrowing nematode came from and how their fields became infested. By reviewing and studying nursery operations, it is clear that at least some of this contamination is occurring with the planting material.

Nematode biology

Burrowing nematodes emerge from eggs and mature in 18–20 days[2]. A typical female may produce as many as 100 eggs in her lifetime. The juvenile (young) and adult stages have a spear-like organ called a stylet that is used for feeding.

These nematodes are obligate pathogens and must feed on living plants. Thus, when plants are first infected, the abundance of healthy tissue provides an ample source of food for nematode growth and reproduction. As the plant is consumed or rotted, more nematodes are produced and less food is available at that site. Nematodes begin to migrate within and outside the plant. Thus, a root system that is 25–50 percent diseased has more nematodes than one that is 95 percent rotted or dead.

Nematodes migrate at any time, but more of them migrate as the root system is rotted and the food supply is decreased. In citrus fields, *R. similis* survives less than 6 months without the host. However, presence of weeds or crop hosts in or near citrus fields allows the nematode population to persist for longer periods. In hot, dry soil, most nematodes will be dead in 3 months, but they may survive for 6 months in moist, cool soil.

In nature, populations of plant pathogenic nematodes such as *Radopholus* are reduced by various means including nematode-trapping fungi, predatory arthropods, oligochaetes, other nematodes that feed on nematodes, bacteria that infect nematodes, starvation, and desiccation. The burrowing nematode produces enzymes that allow it to feed on living anthurium cells, but it cannot feed on dead plants or survive for long periods outside the host. Volcanic cinders provide good drainage, aeration, and contain few, if any, fungal pathogens of anthurium. In laboratory isolations, nematode-trapping fungi are commonly recovered from anthurium plants infected with burrowing nematodes grown in cinders. Thus to some degree beneficial fungi may be reducing the burrowing nematode populations in cinder. In addition, comparisons of potting media have shown that anthurium grown in cinder alone have the lowest number of nematodes compared to plants grown in potting mixes using pine-bark compost and cinder; rockwool, cinder, and peat; or cinder and peat[5].

Disease prevention and control

Treating established fields

For years the anthurium industry has used fenamiphos (Nemacur®) to control the burrowing nematode. Applied to declining fields, yields are improved within 12 months, with 50 percent more flowers produced[1]. However, this represents a continuous cost for the grower, and loss of this nematicide will occur within a few years due to its de-registration. DiTea®, from Valant Biosciences, provides control but requires highly specific site application and management. Testing of other new compounds has revealed that few good possibilities for chemical control of nematodes are likely to become available in the near future.

Starting new fields

Field preparation

New fields should be free of all old anthurium roots or plant tissue, and any cinder applied should be new and unused. Dead plants and roots can harbor a few nematodes that will be the source of inoculum for the next crop. In the citrus industry, the nematodes follow the roots deep into the ground and are present as much as 13 feet (4 meters) deep in the soil. If deep cinder layers are used, deep roots must be removed. Any site that has been used to cultivate anthurium must be scrupulously and meticulously cleaned, or contamination of the new planting will occur. Cleaning the entire field and leaving it in fallow without weeds for at least 6 months is another possibility. Fumigants such as Vapam® can also be used, but efficacy will depend on proper application of the fumigant.

Nursery design

The general nursery design should address the means by which nematodes spread. These include re-use of cinders, water movement, transport and handling of media and plants, use of tools, and traffic by carts and other vehicles to apply fertilizer, spray pesticides, or harvest flowers.

Irrigation

Greenhouse drainage systems should be designed to avoid surface movement of water from one production area to another. Beds should drain into aisles that drain outside of the greenhouse, and each greenhouse should drain to an external, non-production area. It is desirable to plan for prolonged rains that could flood sections of the greenhouse. When planting a field with a slope, plant the lower sections first and proceed to the top to minimize movement of nematodes in older plantings to newer fields.

Employee and visitor movement

Plan walkways and employee movement so there is minimal transport of soil and cinders from an older field through a newer planting. Avoid moving soil or cinders adhering to footwear or clothing. An option is to have a pathway around the perimeter of the nursery for employee movement.

Operations

Avoid nursery operations such as cleaning diseased fields and planting new fields in the same day. Reverse these activities, or use different employees. Gloves, aprons, tools, and other things used in diseased fields should not be used in new, clean fields. If they are used for both fields, then all items must be disinfested by washing with detergent and bleach. The disinfesting solution should be 10–20 percent bleach (1 cup of bleach to 9 cups of water for 10 percent; 2 cups of bleach to 8 cups of water for 20 percent). Metal tools such as clippers can be quickly dipped in chlorine disinfesting solution and rinsed immediately, or alcohol or another surface-sterilizing material can be used.

Cinders

Cinders in fields infested with burrowing nematode should be bare-fallowed or fumigated. For new fields and potted plants, only clean, new cinders or potting medium should be used. Cinders from used pots or infested spots in planted areas should be gathered and dumped in an infested field for eventual treatment. Never leave pots—with or without dead plants—or parts of diseased plants piled around the nursery. These are inoculum sources and should be stored in an area known to be contaminated or discarded at the city dump.

Vehicular movement

Review nursery operations to reduce possible contamination of clean areas of the nursery. For example, do carts travel from older diseased fields to new fields to gather the harvest? If so, reverse the procedure.

Tissue-cultured plants

The use of tissue cultured plants generally ensures that new fields are started without the burrowing nematode. The high value of plants grown aseptically in flasks is their uniform genetic character and the fact that they are pathogen-free. Within the “clean area” where plantlets are removed from flasks and planted, only new pots, tags, and potting media should be used. If trays or pots are reused, they must be thoroughly washed, soaked in 20 percent household bleach solution for 10 minutes, rinsed in running water, and dried. Potting media should never be reused.

If tissue-cultured plants are not grown in a separate greenhouse with careful attention to sanitation, they are likely to become contaminated and will be a source of low levels of burrowing nematodes.

Disease-free mother stock

If the cost of tissue-cultured plants is too high for standard cultivars, growers should strongly consider a program to propagate clean, disease-free cuttings. This involves the use of a special greenhouse that has plants especially prepared for clean culture and is strictly maintained for disease-free plant propagation. It is not a greenhouse that is both harvested and also used for new plants. Many growers of floral crops such as carnation, chrysanthemum, and poinsettia use disease-free mother stock. Large investments are often made in the structures that house these precious disease-free plants.

Begin by producing clean, disease-free plants as described below. These plants should be planted in pots and maintained in a separate greenhouse for disease-free mother stock. The only plants in this greenhouse are those produced by the procedure for disease-free plants or tissue-cultured plants. Monitor the plants in this greenhouse daily. The person who waters these plants should be alert for any sign of leaf spots or rots, stubby rots, petiole or stem rots, or other disease symptom. Maintain the plants in this area in large pots (at least 10-inch [25-cm] diameter). If any plants are found with suspicious symptoms, remove those plants immediately.

Greenhouse requirements

Section off part of an existing greenhouse or construct a separate greenhouse for the disease-free mother stock. Use a clean area covered with a deep layer of new cinder or gravel. Wood, PVC tubing, or metal can be used for the greenhouse frame. The roof can be covered with fiberglass, thick plastic, or other solid material. Saran or plastic fiber can be used for the walls, which must extend from the roof to the ground. Do not leave a space between the wall and the ground, as this will allow snails, slugs, insects, and rodents access to the greenhouse. Weed mats to cover the entire ground are ideal if affordable.

Benches are required and should be made as economically as possible. Bench height should be at least 18 inches from the ground. Construct new benches and avoid bringing old benches into a new greenhouse. If sturdy older benches are available, remove all plants and scrub to remove all roots, moss, and algae. Dry them completely if possible. Drench with 30 percent bleach solution (3 cups bleach plus 7 cups of water). Turn them over and drench the undersurface as well. Leave the bleach solution on for 3–4 hours and then rinse off. These efforts are needed to prevent the immediate contamination of the disease-free greenhouse with pathogens, algae, and moss. Fungal and bacterial pathogens are easily harbored on wooden benches. Previously used concrete tile blocks must be similarly cleaned and disinfested.

Treat the ground to prevent weed growth and maintain an environment free of weeds, insects, slugs, and snails.

Apply water to the plant roots only and avoid watering any non-planted area to reduce moisture in the greenhouse. Eliminate algae, lichen, and moss as they develop. Never allow the nozzle of the watering hose to touch the ground. Hang the hose to prevent contamination of the nozzle, and use good quality water (that is, not from a lake, stream, or river). If surface water is used, it must be chlorinated to eliminate common water-borne pathogens. These are the minimum requirements for a small, special greenhouse to grow disease-free plants.

Preparing clean cuttings

Obtain cuttings from a field without bacterial blight. Select only healthy, vigorous plants and collect only tip cuttings. A young field with vigorous plants is ideal.

Carefully examine every cutting for any symptom of disease. Remove all flowers, buds, dried sheaths, and lower petioles from the cuttings. Remove unopened leaves and trim mature leaves to remove half or more of the lamina. Flower buds and young leaves will deplete the nutrients in the stem and weaken the cutting.

To establish cuttings, root formation must be given the highest priority. Mature leaves will continue to photosynthesize if light and moisture are provided. These leaves are valuable, but water is lost through leaf stomates, and leaf area should be reduced by trimming. At least a third of the leaf blade can be cut off with a clean clipper for large leaves. Tissue-cultured plants also can be used to establish the disease-free stock.

Prepare a disinfesting solution using 1 cup of household bleach and 9 cups of clean water. Add a few drops of liquid detergent to increase surface contact. Place the cuttings in the disinfesting solution for about 30 seconds to 1 minute. Soak for 2 minutes if the cuttings were gathered from older fields. The chlorine in the bleach kills microorganisms on the surface of the plant, but it will also kill anthurium plant cells. A short exposure time will generally kill tiny microbes with minimal damage to the plant. However, exposed young tissues such as root tips may be damaged. The disinfestation treatment only kills the microbes on the surface of the cutting that come into contact with the solution. Nematodes on the inside of the plant will not be affected, which is why the selection of symptom-free cuttings is so important.

Place the cuttings from the disinfestation dip on clean newspaper and plant them after a few minutes. If the cuttings are left overnight, cover them with a moist paper towel or newspaper. Never leave cuttings unplanted for more than a day. Plant the cuttings, one per pot, in 4–6 inch pots. Use Pro-mix™, vermiculite, Sunshine™ blends, or other commercial potting mixes. Consult with UH-CTAHR Cooperative Extension Service agents and other growers to select a high-quality mix. Do not use compost mixes.

Keep cuttings well watered and fertilized to encourage rapid root development. After 2–3 months, carefully remove the plants from their pots and check the roots. All roots should be white, without spots or rots. Immediately discard any plants with brown roots. Repot plants with healthy roots in larger pots using clean cinders or potting mix. For every 100 cuttings, if more than 5–10 plants have brown roots, check roots again after a month. Review nursery procedures with the CES agent to determine why plants are not clean. As with tissue-cultured plants, use only new pots and trays and never reuse media. After the plants are established, the top of each plant can be cut to make another clean stock plant. New shoots can be used for new fields or to increase the mother stock.

Using clean cuttings without establishing a disease-free mother stock

For growers that cannot establish a disease-free mother stock, the following are options to use. Follow the process for producing clean cuttings. Grow the trimmed, disinfested cuttings in pots in a clean area of a saran or plastic-fiber covered house on benches. After the cuttings are rooted they are ready to be transplanted into the new field. At this time, carefully check the roots as the plants are unpotted in the work shed. Do not unpot the plants in the new field. If any plant has root rots, root lesions (long, brown spots), or stubby roots, immediately discard the plant and all its medium. Place the pot in a special box for contaminated pots, or discard it. Use only plants with completely clean roots for the new field.

If gobos are used, collect cuttings only from healthy plants. Do not use plants with stubby roots. Carefully scrutinize the plants for pin-point spots inside the stem. Check for petiole rots and any other symptom of nematode infection or infestation. Place the gobos in a hot-water bath at 120°F (49°C) for 10 minutes(3). For very thick stems, use 122°F (50°C). Place heated, treated gobos on clean newspaper to cool, and then plant them as soon as possible. This should eradicate low populations of nematodes inside the stem, even when visual symptoms may have been missed. Plant the gobos one per pot using new pots and potting medium, and monitor them weekly for any sign of root or stem rots. New shoots should have white roots, and roots produced by the gobo should be white and disease free. Discard the entire pot if any root rots develop.

Employee education

Employee education is crucial to disease prevention and control. Meet with employees to discuss symptoms and signs of nematode diseases, movement of nematodes, known areas of nematode infestation in the field or nursery, and all the efforts being made to control the problem. If a disease-free mother stock is to be established, review the procedures for maintaining this healthy plant stock. Discuss the role each employee will play in the development of all disease prevention and control practices. CTAHR extension agents and researchers can be contacted to conduct seminars and workshops for employee education.

References

1. Aragaki, M., W.J. Apt, R.K. Kunimoto, W.H. Ko, and J.Y. Uchida. 1984. Nature and control of anthurium decline. Plant Disease 68:509–511.
2. Shurtleff, M. and C. Averre III. 2000. Diagnosing plant diseases caused by nematodes. APS Press, St. Paul, Minnesota.
3. Sipes, B.S., D.P. Schmitt, and S.C. Nelson. 2001. Burrowing nematode, a major pest in the tropics. University of Hawaii, CTAHR Plant Disease publication PD-21.
4. Uchida, J.Y., D. Ogata, and N. Nagata. 1999. Tomato spotted wilt virus on anthurium. CTAHR Plant Disease publication PD-17.
5. Wang, K-H., B.S. Sipes, and A.R. Kuehnle. 1997. Effect of soilless media on the growth of *Anthurium andraeanum* infected by *Radopholus similis*. Nematropica 27:77–84.
6. Wang, K-H., B.S. Sipes, and A.R. Kuehnle. 1999. *Radopholus similis* in *Anthurium* shoot tissue. HortScience 34:296–297.

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